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Front Cover: *D. intermedia* on the left, *D. intermedia* x *capillaris* on the right, cultivated by Phil Sheridan. Photograph by Robert Gibson. See article on page 52.

Back Cover: A recent *Nepenthes northiana* pitcher (25 cm in height) from the author's largest plant. See article on page 49.

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ICPS, Inc.
PMB 330
3310 East Yorba Linda Blvd.
Fullerton, CA 92831-1709, USA
icps@carnivorousplants.org

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Editors:
Barry A. Rice, P.O. Box 72741, Davis, CA 95617, USA, email: barry@carnivorousplants.org
Jan Schlauer, Zwischenstr. 11, D-60594 Frankfurt, Germany, email: jan@carnivorousplants.org
Page Layout: Steve Baker, email: steve@carnivorousplants.org

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Contents

Seedbank ----- 35
Genlisea Traps—A New Piece of Knowledge ----- 36
Further Problem in *Genlisea* Trap Untangled? ----- 40
Zero water flows in the carnivorous genus *Genlisea* ----- 46
Looking Back: CPN 25 Years Ago ----- 48
Cultivation of *Nepenthes northiana* ----- 49
Curious Natural Hybrid Sundews ----- 52
Observations on Life Strategies of *Genlisea*, *Heliamphora,* and *Utricularia* in Natural Habitats ----- 57
Literature Reviews ----- 61
Names of Cultivars Registered in 2002 ----- 62
Book Review ----- 63

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Genlisea Traps—A New Piece of Knowledge

Miloslav Studnicka • Botanic Gardens Liberec • 460 01 Liberec • Czech Republic • botangarden@volny.cz

Keywords: observations: *Genlisea, Utricularia* — physiology: *Genlisea, Utricularia*.

Introduction

The subterranean traps of *Genlisea* have been described in detail by various authors such as Lloyd (1942), Fromm-Trinta (1979), Slack (1979), and others, but while these workers examined the details of plant morphology, anatomy and function, they did not answer an important question: What attracts prey organisms into *Genlisea* traps? Slack (1979) speculated on this topic, "It is not clear whether the trap has any particular power to attract its prey, however. The outer parts are scattered with stalkless (sessile) glands which secrete mucilage, but it is not known whether this is of an alluring nature." Meanwhile, recent authors speculated that *Genlisea* traps could function via suction, by expelling water from the trap interiors (Juniper, Robins and Joel, 1989: 87; Meyers-Rice, 1994), but see Studnicka (1996). Recent studies by Barthlott *et al.* (1998) indicate that microfauna such as protozoans may be attracted to *Genlisea* traps. Still, the question remains—what is the attracting agent?

I would like to present a novel explanation that might explain how *Genlisea* traps attract prey to their interiors.

The Trap as a False Soil Interspace

Observing subterranean organs of *Genlisea* we find only a portion of the system of traps in the aerated upper soil layer. A conspicuous number of traps, which are sometimes rather different from the first, are positively geotropic and reach lower parts of the soil profile (Figure 1). In wetlands these parts of the soil can be anaerobic—such conditions are inhospitable to oxygen-dependent microfauna. Oxygen transfer through the body surface becomes inefficient for such organisms in anaerobic conditions (Rusek, 1998). This limitation is especially true at higher temperatures, because of the very low oxygen content in warm water.

Since oxygen availability is a limiting factor for soil microfauna, they are attracted to the tiny air cavities found between soil particles (Rusek, 2000). It is in these oxygen-starved regions that the descending type of *Genlisea* traps are found. If there is an air space within the traps, they should be the best and universal lure for prey in anaerobic conditions. *Genlisea* may be attracting prey by providing an oxygen-rich, false soil interspace.

An Experiment

If *Genlisea* traps were to act as false soil interspaces, we would expect that the neck of the trap might not merely be a tube filled with water. Rather, we would expect the tube to be a more complicated structure in which air cavities would occur—indeed, the trap interior is complex, and adorned with many bristles and cavities.

Figure 1: The traps of *Genlisea hispidula* reach to anaerobic soil space, because the pot was nearly completely immersed in water.

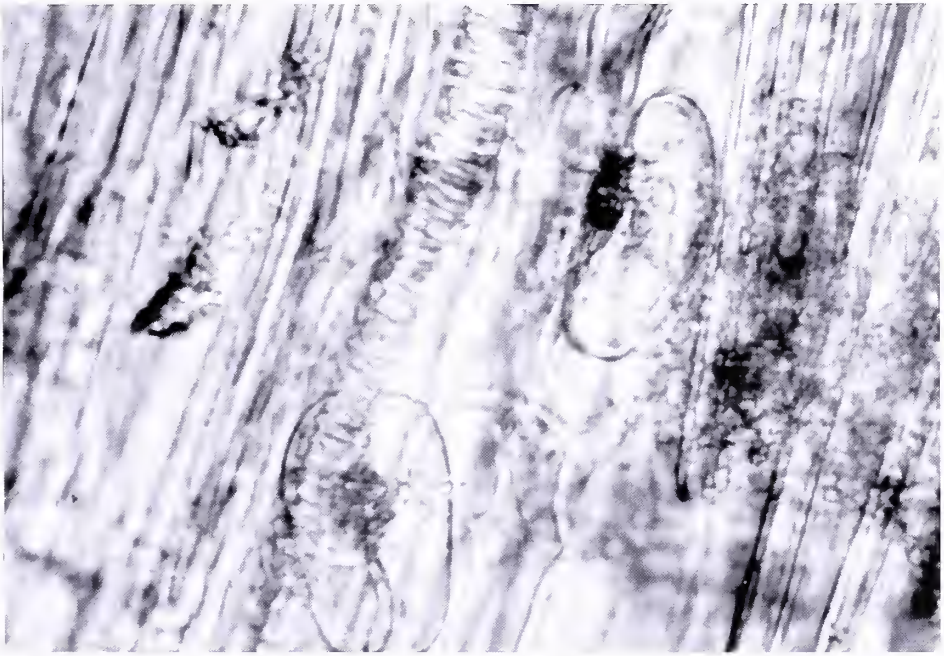

Figure 2: Inner glands in the neck of a *Genlisea* trap (seen from above and in profile). The 2-celled gland head (looking much like a coffee grain) is sitting on the eccentric endodermal cell.

Figure 3: A portion of the interior of a *Utricularia reniformis* trap, near the trap entrance. The 2-celled glands could be analogous to the glands seen in the Figure 2.

To investigate the nature of *Genlisea* traps, a complete trap of *Genlisea hispidula* was transferred from its soil medium to clean water. The forked portion of the trap was removed and discarded. Holding the trap with needle-tipped forceps near the utricle, a second set of forceps was used to squeeze the contents out of the trap-tube (like toothpaste out of a tube). Even though the trap was held underwater for this entire procedure, air bubbles were clearly observed being forced out of the trap.

I also confirmed that the tissues of the *Genlisea* trap are well aerated by lysigenous intercellulars (so called aerenchyma), as reported by Lloyd (1942). Aeration of such underground organs in amphibious plants is a common phenomenon (Votrubová & Soukup, 1999).

Discussion

I conclude that both air and water exist inside *Genlisea* traps. The air cavities are possibly maintained separate from the water by the rows of bristles in the trap neck. This is analogous to the trichomes in *Salvinia* leaves.

We can go back to the question regarding whether *Genlisea* traps are passive or active. The conjectures of Juniper *et al.* (1989) and also Meyers-Rice (1994) may be true in the following special sense! The 2-celled glands in the trap necks (see Figure 2; also see Lloyd, 1942; Fromm-Trinta, 1979) should be compared to the bifid glands in *Utricularia* traps (Figure 3). Inside *Utricularia* bladders, near the trap mouth, there is an area covered by bifid glands which is probably important in exhausting water from the bladders (Sydenham & Findlay, 1975). Perhaps the glands in the neck of *Genlisea* traps, situated between the trap wall and the inner layers of bristles (being covered by bristles) also pump water out of the neck. It would be helpful for air to pass through the plant aerenchyma into the neck.

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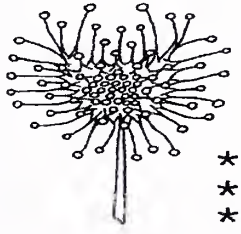

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Further Problem in *Genlisea* Trap Untangled?

Miloslav Studnicka • Botanic Gardens Liberec • 460 01 Liberec • Czech Republic • botangarden@volny.cz

Keywords: carnivory: *Genlisea hispidula* — observations: *Genlisea hispidula.*

Introduction

It is well known that plants in the genus *Genlisea* produce subterranean, Y-shaped, carnivorous trapping organs. The two branches of each trap function as unidirectional channels, directing prey towards the trap neck. At that point, prey travel along the trap neck (a long tunnel) to the vesicle, which is the enlarged digestion chamber.

While these aspects of *Genlisea* are well know, many aspects of how the traps work are still mysteries. Current research is focused on using physiological experiments to test several hypotheses. For example, it is being investigated which (if any) chemical substance attracts prey into the traps (Studnicka, 2002). There are even different opinions as to what kind of prey *Genlisea* tends to capture most frequently. Recent study suggests that *Genlisea* may be specialized to capture protozoa (Barthlott *et al.*, 1998), while Lloyd (1942, p. 94) wrote: "The captures consist of copepods, and the like, small water spiders, nematodes and plenty of other forms, many of which I have seen in the Brazilian material studied." I have observed in *G. pygmaea* many prey organisms, i.e. nematodes, remains of arthropods, etc. (Studnicka, 1996). Furthermore, while preparing the photographs of *G. violacea* glands for this article, I observed numerous nematodes as well as mobile, unicellular organisms in the traps. Some *Genlisea* species produce traps of two distinctly different sizes; this seems to imply some kind of prey specialization that differs between the trap types (Studnicka, 1996).

Another interesting question is how the *Genlisea* traps retain the nutrient molecules obtained by digesting prey items. Since the trap apparently remains open, such nutrient molecules might diffuse out the trap neck and would be lost to the plant. Meyers-Rice (1994) presented a mathematical argument suggesting that traps were active, i.e constantly pumping water through vesicle glands, so the resulting flow through the trap neck would overcome diffusion effects. However, Adamec (2002) found that the flows predicted by this model were not present in *Genlisea* trap necks. In this paper, I present a novel explanation of how *Genlisea* traps may safeguard their precious, prey-derived nutrient molecules.

Hypothesis: A Hitherto Unsuspected Valve?

The tunnel-shaped traps of *Genlisea* are situated in soggy soil and are thus submerged in water. The prey in the vesicle is processed and substances from it are dissolved. Since there is an uninterrupted, free path of water from the vesicle to the trap exterior (i.e. through the open tube to the trap neck), nutrients released by digestion processes in the vesicle could diffuse through the neck and be lost lost to the plant. Careful descriptions of the trap (i.e. Lloyd (1942)) have demonstrated that there are no plant structures in place that would prevent such nutrient losses.

Since there are no structures (based upon plant tissues) that block nutrient

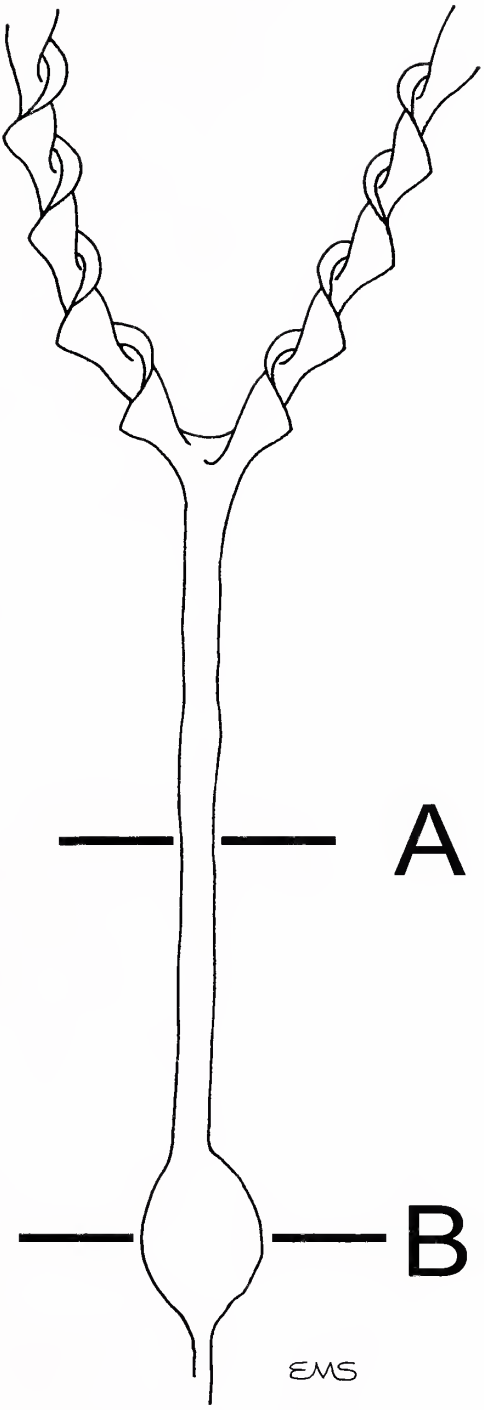

Figure 1: Diagram showing the locations of the slices in the comparisons of the traps to a glass capillary tube. Drawn by E. M. Salvia.

losses from the traps, I have hypothesized that there could be some other kind of barrier or valve holding the dissolved nutrients near the digesting glands. This paper describes laboratory tests and new observations on the nature of internal trap glands which support this hypothesis.

Furthermore, I have found it useful to divide the trap neck (i.e. the trap tunnel that extends from the vesicle to the trap bifurcation) into two portions: the proximal neck (the tunnel near the vesicle) and the distal neck (the part of the tunnel that extends from the trap bifurcation to the proximal neck area). Previous researchers have not recognized the value of dividing the trap neck into two such zones.

Material and Methods

I used *G. hispidula* in the physiological experiments since it produces the largest traps of all the plants in our greenhouse collection. If the trap neck were truly an uninterrupted, fluid filled, hollow tube, it could be considered a capillary tube. As such, it should be similar in many ways to a simple glass capillary tube.

For my first experiment, I used a syringe to pierce the vesicle of an intact trap, and injected a small volume (approximately 0.5 mm^3) of very dilute gentian anilin (known also as methylrosanilini chlorati, approximately 40 ppm dilution). Even at such low concentrations, the purple gentian anilin was clearly visible to the unaided eye. The diffusion of the dye was monitored.

In my second experiment, I prepared a small frame to hold both glass capillary tubes and *Genlisea* traps in a vertical orientation, above a glass Petri bowl filled with water. I prepared a glass capillary tube with the same approximate inner diameter of a *Genlisea* trap neck (i.e. approximately 0.2 mm). I also prepared two *G. hispidula* trap necks. On "Trap A" I removed the vesicle and proximal neck (i.e. the cut was made through the central neck portion), while on "Trap B" I cut through the vesicle (see Figure 1). The glass capillary tube and the prepared trap fragments (oriented cut-side downward) were mounted on the frame, and were then lowered so the ends of all three contacted the water in the glass Petri bowl. Water was immediately drawn into the tubes by capillary action, so the three tubes could be considered similar, water-filled capillary tubes. Subsequently, several drops of violet anilin solution were added to the water in the Petri bowl.

Surprising Results

In the first experiment (where purple anilin dye was injected into the *Genlisea* vesicle), the colouring did not spread beyond the proximal neck area (see Figure 2). Even after an hour had passed, the anilin dye did not spread beyond the proximal neck area. This experimental result was the same whether the trap was submerged in water or kept in the air.

In the second experiment, involving the glass capillary tube, *G. hispidula* Trap A, and *G. hispidula* Trap B, there was a very interesting result. The glass capillary rapidly became coloured as the dye solution diffused into the water column. Similarly, in Trap A, the anilin quickly diffused throughout the distal neck (see Figure 3). In contrast, in Trap B, the dye was unable to diffuse out of the vesicle and proximal neck area!

Gland Observations

The trap neck has numerous glands on the internal tube surface. The glands of *G. violacea* were studied by coloring them with iodine tincture, which stains cellulose in particular. The heads of the glands in the proximal neck area of the *Genlisea* trap are multicellular (see Figure 4). Searching for similar glands in related genera, we find near-

Figure 2: The *Genlisea hispidula* vesicle and proximal neck area after the injection of purple anilin into the trap vesicle. The dye cannot diffuse into the central neck area.

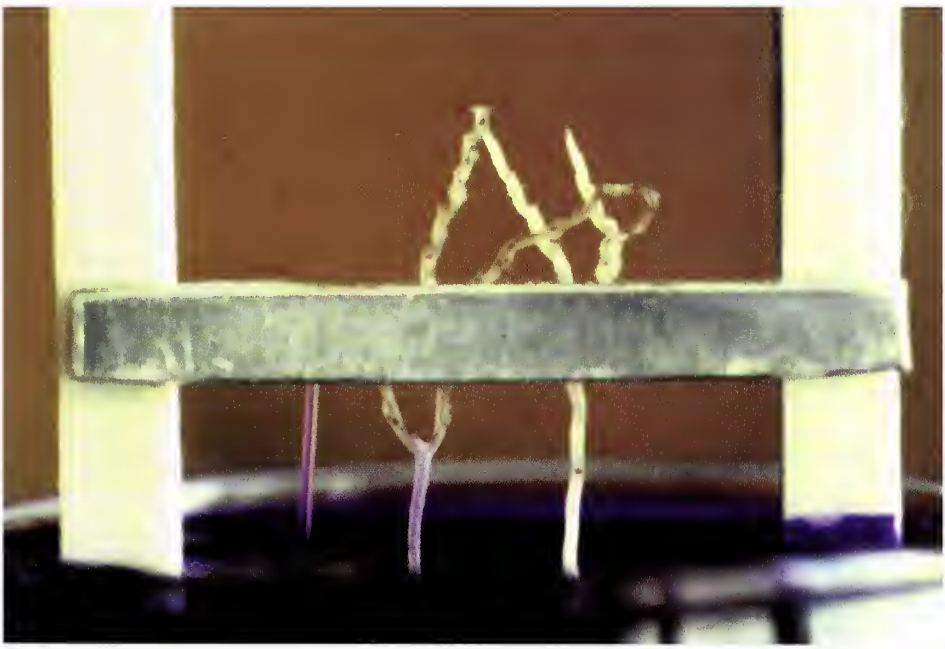

Figure 3: Diffusion of the violet anilin colour into the glass capillary tube (left) and Trap A (center). No diffusion is evident in Trap B (right).

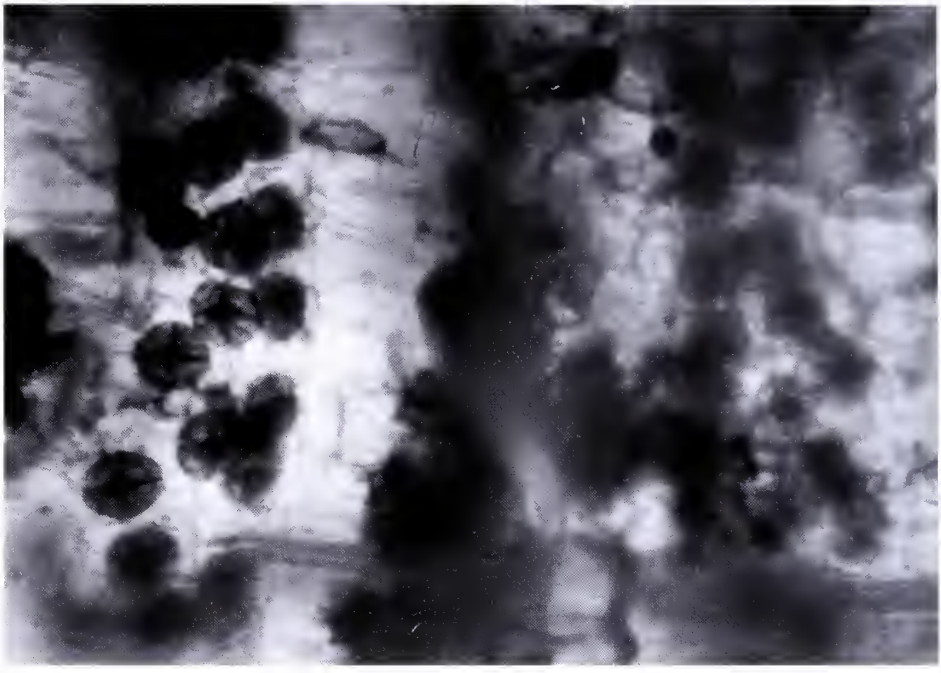

Figure 4: Glands in the proximal part of the *Genlisea violacea* trap neck.

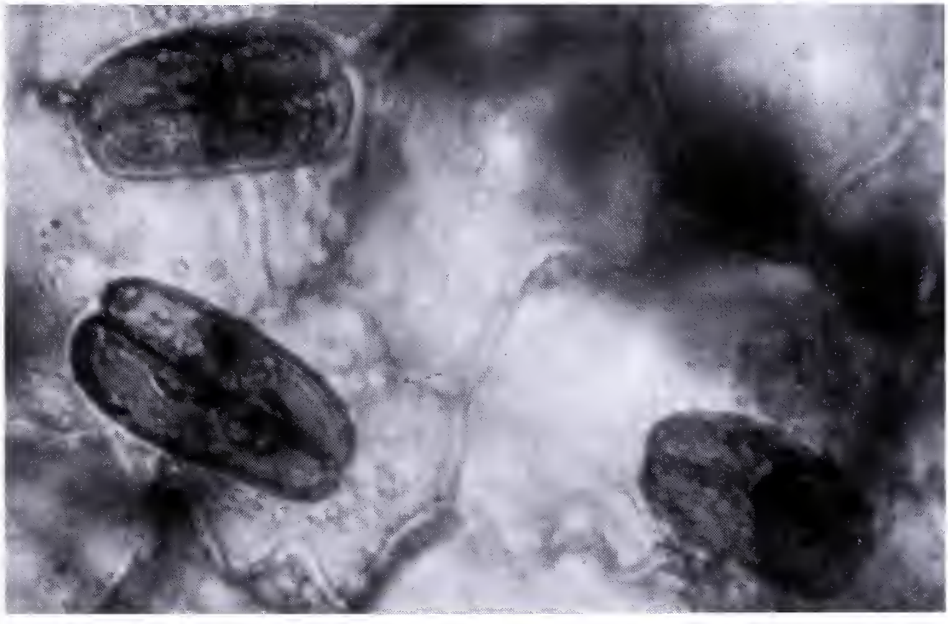

Figure 5: Glands in the distal part of the *Genlisea violacea* trap neck.

ly the same glands in *Pinguicula* (i.e. the sessile digesting glands; see Juniper, Robins & Joel 1989: 153).

In contrast, the heads of the glands in the distal portion of the trap neck are only 2-celled (see Figure 5), and rather resemble the bifid glands found near the mouth of *Utricularia* bladders (Lloyd, 1933: 45; Sydenham & Findlay, 1975: 348).

Interpretation of the Experiments

It seems very likely that the two different gland types have two different functions.

If we fix our attention on the glands in the proximal neck, we may infer that they may serve the same function as the similar glands found in plants in the related genus *Pinguicula*. Specifically, they may secrete a viscous compound which is not too easily miscible with water. (Indeed, *Genlisea* are well known to secrete mucous in other contexts, i.e. from the photosynthetic leaves, e.g. in *Genlisea aurea* as reported by Rivadavia (2002)). It is possible that such secretions would partially or even completely block the capillary-like trap tunnel. If this is indeed the case, nutrients dissolved in the vesicle would be prevented from diffusing from the vesicle and proximal neck into the distal neck (and, subsequently, escaping into the environment). On the other hand, an agile little animal could penetrate this viscous area by means of its own force. The secretion functions similarly to a semi-permeable membrane.

The inability of the anilin dye to diffuse from the vesicle and proximal neck areas into the distal neck areas (as shown by my experiments) supports this interpretation.

The presence of similar glands in both the vesicle and the proximal neck area in the *Genlisea* traps suggest a shared nature—indeed, the proximal neck may be an extension of the vesicle

It is still unclear what the function might be for the glands in the distal neck areas.

Acknowledgement

I would like to thank Barry Rice for reviewing and improving both this and my previous *Genlisea* articles that appeared in Carnivorous Plant Newsletter.

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ZERO WATER FLOWS IN THE CARNIVOROUS GENUS *GENLISEA*

LUBOMIR ADAMEC • Institute of Botany • Academy of Sciences of the Czech Republic • Dukelská 135 • CZ-379 82 Trebon • Czech Republic • adamec@butbn.cas.cz

Keywords: carnivory, physiology: *Genlisea violacea, G. hispidula*.

In carnivorous plants, researchers seek to elucidate unique processes such as enzyme secretion, nutrient absorption from prey, rapid trap movements, and the ecological importance of carnivory. Among these processes, one of the most mysterious is water flow in *Utricularia* (bladderwort) traps. These 1-4 mm long, bladder-shaped suction traps pump out water to maintain internal underpressure (Juniper *et al.*, 1989). After irritation by prey, the trap is quickly opened and the prey is sucked into the trap. No lesser mystery is the functioning of *Genlisea* traps (Juniper *et al.*, 1989; Meyers-Rice, 1994; Studnicka, 1996; Barthlott *et al.*, 1998). *Genlisea* species—wetland or amphibious plants from South America and tropical Africa—are closely related to *Utricularia* (Lentibulariaceae).

Genlisea forms short stems with a rosette of small photosynthetic, green leaves and subterranean white traps. These traps are highly modified carnivorous leaves without chloroplasts (see Figure 1; also Juniper *et al.*, 1989; Studnicka, 1996; Barthlott *et al.*, 1998). The inverted Y-shaped traps are 2-15 cm long and about 1 mm thick. The hollow arms of the trap open into the hollow tubular neck (inner diameter 200-500 micrometers), which ends in a slightly dilated vesicular cavity. The whole central cavity of the arms and neck is filled with water and is lined with detentive hairs pointing towards the vesicle. As such, *Genlisea* traps function as "eel traps." Thus, microscopic prey are attracted to enter the traps and, thanks to the hairs, are led forward to the vesicle. Here, enzymes are secreted, and the prey is digested. It has been found recently that the traps of *Genlisea* are specialized for attracting and catching protozoa (Barthlott *et al.*, 1998). However, besides living or decomposed prey (Barthlott *et al.*, 1998) and sometimes also algae (Studnicka, 1996), fine soil particles have often been found in the vesicles (Juniper *et al.*, 1989; Meyers-Rice, 1994; Studnicka, 1996).

Based on this finding, on overall similarity to traps of *Utricularia*, and on considerations of the absorption efficiency of the traps, a hypothesis of active water flow in *Genlisea* traps exists (Juniper *et al.*, 1989; Meyers-Rice, 1994) with a supporting mathematical model (Meyers-Rice, 1994). Assuming the same water flow rate in *Genlisea* traps per trap wall unit area as in *Utricularia* traps during the resetting phase, a water flow rate of 14 micrometers per second in the neck cavity towards the vesicle was calculated. This rate exceeded by about three-fold a calculated rate of counter-diffusion of phosphate molecules (4.8 micrometers per second) at which phosphate, as a typical product of digestion of prey, could diffuse back from the vesicle and be lost from the trap. If this were so, the active permanent water flow would drag suspended soil particles and, to some extent, also living prey to the vesicle and prevent nutrients released in the vesicle from escaping due to diffusion (Meyers-Rice, 1994).

To test this hypothesis, I measured water flow in large traps of *G. violacea* (a "giant form" in horticulture) and *G. hispidula*. The plants were grown in a pot in a mixture of filamentous peat and perlite in a naturally lit greenhouse. Water flow through the traps was measured using a capillary glass microvolumeter (Dainty & Ginzburg, 1964) with a resolution of approximately 0.05 microliters and stability

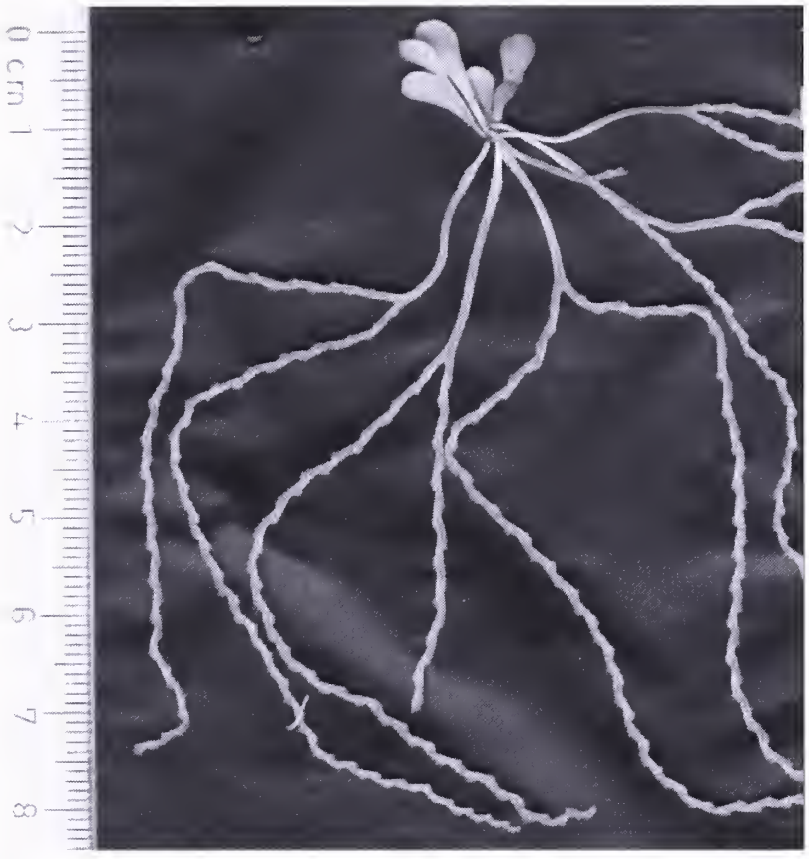

Figure 1: *Genlisea violacea* plants with green leaves and apoplastic white traps. The upper dilated part of the neck is the vesicle.

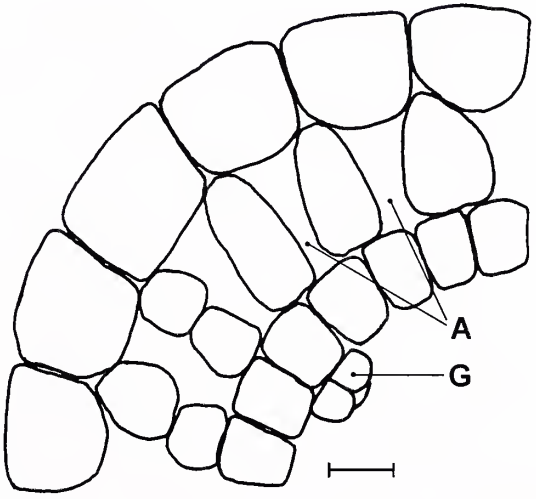

Figure 2: Schematized cross-section through *Genlisea violacea* vesicle. A: air spaces; G: sessile gland. The bar represents 50 micrometers.

better than 0.03 nl s^{-1}. The expected water flow could range within approximately 0.17-3 nl s^{-1} (Meyers-Rice, 1994). Traps of experimental plants were carefully released from the substrate and washed using tap water. A trap with shortened arms and without leaves was fixed using silicon cement in the neck of this device just above the bifurcation of the trap. After temperature stabilization in a thermostatted bath (22.0±0.02°C) in darkness, water flow was measured for 40-60 min.

Counter to the model, virtually zero water flow was measured in both species (n=8-10 in each species). These measurements prove that *Genlisea* traps are passive. The soil particles often observed in the vesicles could enter by another route, e.g. if pushed by captured prey (Studnicka, 1996). In light of these negative experimental results and the anatomy of the vesicles (see Figure 2), it is possible to offer several other arguments against the hypothesis of active water flow in *Genlisea* traps. *Genlisea* traps lack bifid glands which would be responsible for water pumping as in *Utricularia* (Juniper *et al.,* 1989; Studnicka, 1996). The wall in *Genlisea* vesicles consists of 3-4 layers of large cells with voluminous air spaces, unlike the mere two layers of cells, without air spaces, in *Utricularia* traps (Juniper *et al.* 1989). The presence of large cells and voluminous air spaces would slow down any osmotically driven water flow. *Utricularia* traps pump water for only 20 minutes until the traps are ready to fire (Juniper *et al.,* 1989; Meyers-Rice, 1994). The always-open *Genlisea* traps would have to pump water constantly to prevent loss of nutrients. Accordingly, such an operation would demand a large amount of energy, and the energetic cost would probably exceed the nutritional benefit. However, *Genlisea* traps might efficiently use mineral and organic nutrients released from prey in the vesicle, even without pumping water, if the rate of absorption of these nutrients in the vesicle and neck is sufficiently high. Merely a high density of absorptive sessile glands in the vesicle and along the neck could give evidence for this hypothesis.

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Looking Back: CPN 25 years ago

"In many plants, exsiccata (dried herbarium specimens) have the disadvantage of a considerable information loss. In such cases color photography may offer a valuable additional source of scientific information." With these sentences, Jurg Steiger began an eight page article about recording *Pinguicula* floral variations by standardized photography. This article is a gold mine of information that includes 27 photographs of 81 *Pinguicula* flowers of many species. If you never bought this back issue when the ICPS was selling them, lament!

Cultivation of *Nepenthes northiana*

Dr. Jeff Shafer • neps@plantswithattitude.com

Keywords: cultivation: *Nepenthes northiana.*

One of the most beautiful of all lowland nepenthes, *N. northiana* hails from the limestone hills of the Bau region of Sarawak, Malaysia. Although most lowland *Nepenthes* are considered easy to grow (at least relative to the more slowly growing highland species), *N. northiana* has acquired a reputation for difficulty in cultivation. Having grown this species for several years now with some success, I feel qualified to share my observations and cultivation techniques with other growers, and hope to dispel some of the mystery surrounding this remarkable plant.

My experience growing this species in Colorado, USA, began about five years ago, when I received a rather small, bedraggled specimen, approximately 2 cm (less than 1 inch) in diameter. This seedling had been delayed in shipment and arrived in deplorable shape. Fearing that it would not survive, I placed it in compost comprised of fine grade horticultural charcoal and sphagnum peat moss, in the approximate ratio of 3:1. The specimen was then located in a partially closed plastic bag, and situated in a shady spot in my lowland *Nepenthes* greenhouse.

To my delight it survived, and after about two weeks I removed the plastic covering. Initial growth was tentative, until the appearance of pitchers which I began to feed with extremely small crickets. All my *Nepenthes* are hand-fed with insects, a process which is highly time-consuming for a collection of any real size (not to mention distasteful); however, I believe that much of the success I have enjoyed cultivating members of the genus is due to this approach. In any case, within a few weeks of the initial feeding, the rate of growth and size of successive leaves of my specimen began to increase. However, the plant was very slow to develop an extensive root system. In fact, for about a year, it was rooted so tenuously that unless great care was taken while watering, it would become unanchored and drift in its pot.

After about one year, the plant had reached a size of 8 cm (3 inches) or so, and it was at this point that it finally began to settle down and produce a reasonable root system. As chance would have it, my plant had settled near the edge of its pot, and its roots had grown down through an area of almost pure charcoal, allowing it excellent drainage. This was something I noticed after another year had passed, with the plant now about 15 cm (6 inches) in diameter and in need of placement in a larger pot. At this time, I placed it in a 15 cm vessel and used compost comprised of fine horticultural charcoal, fine orchid bark, and sphagnum peat, in the approximate ratio 3:2:1. I do not think that the use of orchid bark is in any way critical, but I often use it with charcoal to ensure good drainage, and because it is light and relatively inexpensive. No other compounds were added to the compost. I emphasize this point because there has been considerable speculation among several growers with whom I have conversed regarding the need for limestone, antimony, or other such substances in the compost for *N. northiana*, and much of the lack of real success in the cultivation of this species has been anecdotally attributed to the absence of these or other, as yet unknown, substances. I have found no reason to believe that anything other than what I have described need be used as compost for the successful cultivation of *N. northiana*!

Repotting was done in the middle of autumn, a time when temperatures are generally moderate at my location, and my greenhouse environment is typically devoid of extremes of temperature and humidity. And, for about the next six months or so, my *N. northiana* was fine, having settled into its new compost and produced a number of new,

ever larger leaves and pitchers. However, with the advent of spring that year, I began to notice some problems. Specifically, pitcher production ceased, and the leaves of the plant began to exhibit a bronze tint, with circular patches of dead tissue approximately 0.5 to 1.5 cm (0.2-0.6 inches) in diameter. Relocation to a less well-illuminated area mitigated the latter problem, immediately implying that this species may best be situated in a low-light environment. This was confirmed for me by several individuals who have had the good fortune to observe this plant *in situ*, and who have informed me that specimens growing in areas with significant direct sunlight tend to be less healthy and robust than those better shaded.

However, relocation of the plant to a less well-lit location did not result in the production of new pitchers. Furthermore, since the absence of pitchers had preceded the relocation of the plant, I did not think it likely that reduction of illumination was the cause. In fact, the crux of the problem was the relative humidity of the area in which I live. Typically, our humidity can drop into the teens (sometimes even into single digits—yikes!) during the summer months. (Temperatures in the growing area are approximately 28-36°C, 82-97°F during the day, and 20-24°C, 68-75°F during the night.) The necessary venting of my greenhouse to mitigate solar heating causes pronounced fluctuations in the humidity in the greenhouse. To combat these problems, I installed humidification equipment which raised the humidity to about 75%. As a result, all my lowland *Nepenthes* plants pitcher quite regularly—with the exception of *N. northiana*!

So, since my plant was still not pitchering well, I decided to place it in a lowland environment with stable humidity. To do this, I put it into an aquarium with a thin sheet of clear Plexiglas covering the top. About two liters of water were poured into the tank to provide ample humidity, and the plant was placed on a stand so that it would be out of the water. Directly above the aquarium, I placed four 40 W fluorescent shop lamps (just standard shop lamps, not the broad-spectrum type typically touted as essential for growing plants) on a timer with a 13 hour photoperiod. The plant was watered thoroughly every two or three days with water purified by a reverse osmosis unit.

Much to my delight, my plant immediately began to pitcher again, but I noticed that the leaves were still developing the now familiar bronze tint. Therefore, I reduced the number of lamps illuminating the enclosure by half, and subsequent growth was a healthy green again, with no cessation of pitcher production! Since that time, the growth of this plant has been uninterrupted, with the size of the specimen increasing dramatically. At the present time, leaves are 65 cm (26 inches) in length, and I have had to construct a custom enclosure to replace the aquarium which it has long since outgrown! Pitcher size has increased to nearly 25 cm (10 inches), which although nowhere near the 40 cm (16 inches) commonly seen in the wild, is still respectable, with the pitchers exhibiting all the characteristics for which the species is so widely known (see Back Cover). Recently, though, my *N. northiana* did stop pitchering, and this was a matter of some concern initially. However, a brief examination of the roots quickly revealed that the plant was root bound. I placed it in a 45 cm (18 inch) pot, and it has now happily resumed its familiar growth.

Having therefore enjoyed a reasonable degree of success growing this beautiful species, some time ago I decided to obtain a couple of new specimens from different sources for further experimentation, making sure that these new acquisitions were not clonally identical to my current specimen in the interest of ensuring that earlier success was not due to the good fortune of receiving a particularly hardy clone. Therefore, I placed one of the new plants in a mix essentially identical to that previously described. In addition, I placed the other plant in compost consisting of almost pure peat moss, in an attempt to determine the extent to which much more absorbent compost might affect growth. I am happy to report that after two years of experimentation, both of these plants are growing and pitchering quite well, with the choice of compost having made little apparent difference!

Thus, although my experiences and observations regarding the cultivation of *N.*

northiana are anecdotal in nature, it is my belief that they may serve to clarify some of the confusion regarding the cultivation of this wonderful species. In summary, I believe that there are two key issues which the grower must address. 1: Provide a source of consistent, low-intensity illumination, which can be provided by something as simple as ordinary 40 W fluorescent shop lamps. Failing this, try placing your plant in a location where it will receive indirect sunlight, adjusting if leaf damage as described above occurs. 2: Ensure that your growing environment provides consistent, high humidity and temperatures in the approximate range of 20-35°C (68-95°F). It is my belief that observation of these points, along with the other general points of good *Nepenthes* horticulture, will likely enable the reader to enjoy the successful cultivation of this most remarkable and beautiful plant!

Those interested in sharing their experiences with this species, or who may have markedly different observations, are encouraged to contact the author.

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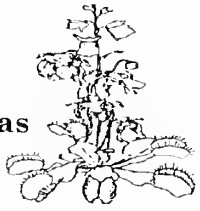

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Curious Natural Hybrid Sundews

Ivan Snyder • 110 Meyer Court • Hermosa Beach, CA 90254-5351 • USA

Keywords: genetics: *Drosera capillaris, D.* × *obovata.*

One Failure, One Success, One Mystery

You may remember me from my earlier colchicine treatment article (Snyder, 2000). Since that article, I have learned about some curiosities along similar lines that I would like to share with you. I would like to focus on wild *Drosera* native to North America.

I wish to discuss how hybridization in populations of plants plays an important role in evolutionary change. By important, I do not necessarily mean beneficial for the survival of the plant populations. We will examine the role of hybridization in three different sundews. The first sundew, *D.* × *obovata*, is an abundant hybrid which has proven to be an evolutionary dead-end. The second sundew, *Drosera anglica*, began as a hybrid and developed into a successful species[1]. The third sundew, *D. capillaris*, exhibits signs of amphiploidy.

North American Species and Hybrids

In North America there are seven species of *Drosera*. These are *D. anglica*, *D. brevifolia*, *D. capillaris*, *D. filiformis*, *D. intermedia*, *D. linearis*, and *D. rotundifolia*. All seven are capable of hybridizing with one-another, giving a total of 21 possible F1 hybrid combinations. But not all 21 will be created naturally since not all seven are within cross-pollination distance. The three most common interspecies hybrids have been formally described and named. The most common natural hybrid by far is *D.* × *obovata* = *anglica* × *rotundifolia*. Next is *D.* × *beleziana* = *intermedia* × *rotundifolia*, followed by *D.* × *hybrida* = *filiformis* × *intermedia*. The latter two may be found in the New Jersey Pine Barrens. From North Carolina is *D. capillaris* × *intermedia* (Sheridan, 1987). One hybrid reportedly seen in Florida is *D. brevifolia* × *capillaris*. I think this one would be especially difficult to identify. Other hybrids are expected to turn up where both parents are sympatric. Two manmade hybrids of sympatric parents, *D. brevifolia* × *rotundifolia* (created by me) and *D. brevifolia* × *intermedia* (created by Jim Bockowski), proved to be good growers, though have not been recorded from nature.

There may be reasons that some hybrids have not been found—reasons beyond their merely having eluded detection (Butler, 1985). I discuss some possible reasons in this paper.

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[1]Note that Art.H.4.1 of the International Code of Botanical Nomenclature requires that the first legitimate name that refers to a taxon of hybrid origin must equally be applied to all subsequent filial generations, back-crosses and combinations of these if all parent taxa are known or can be postulated. So if it is assumed that *Drosera anglica* is derived from a hybrid involving *D. rotundifolia* as a parent species, then the only legitimate name of the back cross *Drosera anglica* × *D. rotundifolia* would also be *Drosera anglica*, and *Drosera* × *obovata* would be a synonym of *Drosera anglica*.—ed. (Jan Schlauer)

Nature's Failure—the Paddle-Leaved Sundew

The familiar *Drosera* × *obovata* (commonly called the paddle-leaved sundew), is a cross between *D. anglica* and *D. rotundifolia*, and was the first sundew recognized to be a natural hybrid. It is found wherever both parent species grow together, and since the very common parents have a vast distribution, surely this hybrid must be the most widespread of all. *Drosera anglica* has long leaves and *D. rotundifolia* has round leaves, so the hybrid has an intermediate shaped leaf which is a long oval or obovate shape, like a canoe paddle. *Drosera* × *obovata* has never been documented to be able to form viable seeds. But is this an unbroken rule for this hybrid? Could this hybrid at times really be fertile?

The reason for infertility is probably, putting it simply, that when two incompatible plants are mated their chromosomes do not match up. Consider the easily understood case of *D.* × *obovata*. The *D. anglica* parent is tetraploid (four sets of chromosomes), and *D. rotundifolia* is a diploid (two sets of chromosomes). The offspring will have one-half of the pair of chromosomes from each parent, making *D.* × *obovata* a triploid (three sets of chromosomes). *Drosera* × *obovata* cannot produce viable pollen (or ovules) because its third set of chromosomes cannot divide or pair correctly.

The first natural hybrid sundew I ever saw was the paddle-leaf, here in my home state of California. I found a fantastic floating bog habitat around the edge of a beautiful lake using a hiking trail book (It is great fun to follow a trail guide, it is like decoding a treasure map). The hike to this habitat just outside the southern boundary of Lassen Volcanic National Park was a most memorable and awesome adventure.

Drosera × *obovata* is very rare in California but can be found in the northern part of the state at high elevation in sphagnum bogs. The treasure my map led me to was growing on rafts of fallen logs in sphagnum moss along with both parent species, and a few *Utricularia* species as well.

Despite my thrill at finding this habitat, I had in mind a search for something even more special. Even more exciting would be to find the most rare of rare; a plant so unknown it has not been formally documented—a fertile paddle-leaf. So while I was treading amongst the great numbers of *D.* × *obovata* growing over a vast area, I examined the plants carefully to see if I could find any that might have seed. (I was there in August at the time when seed capsules on *D. anglica* were fat and full of seed.) It was easy to pick out the flower stalks of the hybrid among the *D. anglica* because the *D.* × *obovata* stalks did not have enlarged seed capsules. Searching through what must have been a million sundews I looked over hundreds of barren stalks. Dissecting several, I found a few underdeveloped seed that I am sure would not have been viable. Still, after studying the matter of fertility in other hybrids, I feel certain there ought to be some that are fertile.

Nature's Success— *Drosera anglica*

As if these glistening carnivorous plants were not already fascinating enough, it seems we have only begun to learn of hybridization amongst the species. This, in my mind, elevates the sundew to a whole new level of fascination. It is a surprise to people the first time they learn that at least one species in North America is actually of hybrid origin. This is *D. anglica*, which started off as a cross between *D. linearis* and *D. rotundifolia* (Wood, 1955). But how can this be fertile? As was told previously, a hybrid with mismatched chromosomes will be sterile. By an accident of cell division a plant's chromosomes can double. This happens when a cell nucleus splits into two without the cell itself dividing. The mismatched chromosomes can then match up with their doubles. Any hybrid that has undergone this process is

Figure 1: A fertile *D.* × *obovata* plant. Photograph by Ed Read.

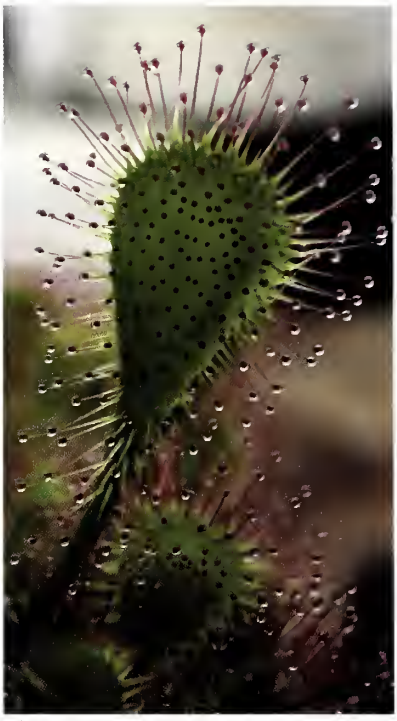

Figure 2: A fertile *D.* × *obovata* plant. Photograph by Ed Read.

Figure 3: The *Drosera capillaris* plant with long leaves. Photograph by William DiLapi.

known as an allopolyploid or an amphiploid. This is a natural process that is not uncommon in plants.

Thanks to the efforts of Donald Schnell, the case of the origin of *D. anglica* is well studied and understood (Schnell, 1999). If what goes on with *D.* × *obovata* is any guide, it seems that countless *D.* × *anglica* must have germinated and died before one became fertile through amphiploidy. This process must have gone on eons for *D. anglica* to become so widespread. Creation of even more fertile *D. anglica* plants may continue where *D. linearis* and *D. rotundifolia* exist together. The origin of *D. anglica* is a good example of how all of nature's creations start off as accidents. While most of nature's experiments are doomed failures or dead ends, and relatively few prove successful.

Amphiploidy may also be induced artificially using chemicals, and can be an important tool in plant breeding. Recently the sundew has become a subject of such tinkering (Snyder, 2000). In theory, all sterile though otherwise healthy hybrids may be made fertile through chromosome doubling, although unpredictable side effects are possible—some plants will have improved vigor, while others are weakened. When the hybrid *D.* × *anglica* is chromosome doubled, the resulting fertile *D. anglica* is a more vigorous plant.

A Fertile *Drosera* × *obovata*?

So far, *D.* × *obovata* seems adversely affected by amphiploidy. When this triploid is doubled to a hexaploid it becomes very weak and unhealthy. This demonstrates that it is probably not capable of becoming a successful allopolyploid in nature. There is still the possibility of a far less common event, the natural creation of a tetraploid *D.* × *obovata*. While a normal triploid *D.* × *obovata* is the result of a cross between a diploid *D. rotundifolia* and a tetraploid *D. anglica*, if the parent *D. rotundifolia* used were a tetraploid the resultant *D.* × *obovata* would be tetraploid. Such a hybrid might be fertile. (Recall also that the *D. anglica* itself is already composed partly of *D. rotundifolia*, so its cross with *D. rotundifolia* will be partly compatible.

Using a tetraploid *D. rotundifolia*, I created a tetraploid *D.* × *obovata* that has proven to be fertile, albeit barely so (Figures 1, 2). (While the tetraploid *D. rotundifolia* has perfectly good pollen, its seed does not completely develop well. I think this flaw is the main reason the tetraploid *D.* × *obovata* has low fertility.) This gives some credence to the unpublished rumors of field discoveries of fertile *D.* × *obovata* plants. Schnell (1999) states that all North American *Drosera* hybrids are sterile, so the discovery of any fertile hybrid plants is important. Considering the vast range in which it could take place, I see no reason why this tetraploid form of *D.* × *obovata* would not also be naturally created, although I suspect this would be an extremely rare occurrence. Sadly, it seems that my prize the fertile paddle-leaf is an evolutionary dead-end. Because of low fertility, it is poorly suited to perpetuate and disperse by seed. This type of hybridization worked for *D.* × *anglica*, but failed in *D.* × *obovata*.

Mystery—A New Species From Florida?

Now let us examine another mysterious plant, something that appears to be a cryptic allopolyploid that, while perhaps fairly abundant in nature, has so far escaped scientific attention. While on a carnivorous plant hunting trip in Florida, fellow carnivorous plant nut Bob McMorris led me to a strange form of *D. capillaris* with very long petioles (Figure 3). (Some horticulturists have grown and traded this plant under the name *Drosera capillaris* "long arm", but this is not a cultivar name.) This plant has long slender upright petioles much like those in *D. intermedia*. Bob had told me of his belief that this plant was a hybrid between *D. capillaris* and *D.*

intermedia. When I first saw specimens, they did indeed appear to be just such a hybrid, but the plants at least appeared fertile because the capsules were full of seed.

At home, I wondered about these plants. Since the seeds germinated very well, it seemed unlikely the plants were a hybrid, and I felt that they were most likely an odd form of *D. capillaris*. After all, this can be a variable species. I have also noticed that the leaf shape is not the only aspect of *D. capillaris* that is variable—the style shape is also variable and the styles on some plants are bifid, much like those of *D. intermedia*. When I noticed this, I decided that this little plant deserves more study. I wondered if hybridization between *D. capillaris* and *D. intermedia* and subsequent allopolypoidy might indeed be occurring.

The idea regarding hybridization was not new with me. Phil Sheridan had reported finding *D. capillaris* × *intermedia* (see Front Cover) growing amongst the parent species in coastal North Carolina, USA (Sheridan, 1987). His article was planned as a preliminary study, but he became involved in other projects and never completed the work. The hybrids Phil found looked much the same as Bob's plants from Florida, but Phil's plants were sterile. Phil told me that he knew of the strange plants in Florida, and also suspected them to be allopolyploids (and possibly a new species).

How could we distinguish whether the plant is an allopolyploid? When a plant's chromosomes are doubled, cell size also doubles. While plant breeders usually compare pollen grain size with a plant of a known ploidy, with sundews I find it easier to simply compare the cells on the leaf surface. This I do with a 20× or 30× hand lens. When I looked at many clones of the strange long-petioled *D. capillaris* from Florida, I was surprised to see that indeed all had cells twice the size of ordinary *D. capillaris*. More testing is needed, but the possibility of a new allopolyploid looks promising.

Keep an Eye Out

We have only begun to unlock the secrets of *Drosera* and hybridization. Strange things lay hidden awaiting discovery. The study of carnivorous plants is unique and can be especially enjoyable. Even an amateur scientist such as myself can make satisfying and important discoveries! If you have the opportunity to visit a bog yourself, keep an eye out for that unusual sundew. Remember, a fertile paddle-leaf is possible, and this hybrid would be an extremely rare prize. There are surely many more sundew curiosities yet to be discovered around the world. Happy hunting!

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Observations on Life Strategies of *Genlisea*, *Heliamphora*, and *Utricularia* in Natural Habitats

Miloslav Studnicka • Botanic Gardens Liberec • 460 01 Liberec • Czech Republic • botangarden@volny.cz

Keywords: observations: *Brocchinia tatei*, *Genlisea roraimensis*, *Heliamphora nutans*, *Utricularia humboldtii*—ecology—carnivory.

In December 2001 I travelled through Venezuela. I expected to visit Mt. Roraima during my travels, and since I was particularly interested in the carnivorous flora in this area, I arrived with as much laboratory equipment as I could bring (Figure 1). I especially hoped that my microscope would let me study the prey captured by *Genlisea*. In this paper, I report on a few novel observations and interpretations that resulted from this trip.

Prey of *Genlisea roraimensis*

In a recent paper, Barthlott *et al.* (1998) presented laboratory observations which suggested that *Genlisea* are specialists in capturing and digesting protozoa. My own observations with cultivated *Genlisea* show they capture many nonprotozoan prey (Studnicka, 1996). This is in agreement with Lloyd, who noted that Brazilian *Genlisea* captured "copepods, and the like, small water spiders, nematodes, and plenty of other forms" (Lloyd, 1942). A weakness of both my research and the work reported by Barthlott *et al.* (1998) was that our research programs both were based on cultivated plants. As such, I was curious to observe what *Genlisea* plants captured in their native habitats in South America.

This interest led me to visit Roraima with a small microscope in my backpack. My microscope can easily detect protozoa in wet soil samples, and I have used it to examine the often-numerous protozoa that occur in the soil of cultivated *Genlisea*. If protozoa were common in either the wild *Genlisea* traps at Roraima or their habitats, I was confident I could detect them easily. During my investigations, my two assistants and I made three interesting observations. First, when we looked for protozoa in association with the *Genlisea*, none were observed within the traps. Second, we found that protozoa were very rare in the moist soil where *G. roraimensis* grew. Since I have used this equipment to detect protozoans in other wet soil samples, I can confidently conclude that the soil we studied on Roraima had very low populations of protozoa. Third, we did observe living, mobile nematodes within the *Genlisea* trap vesicles.

I believe the *Genlisea* traps are indeed well-deserving of the name "eel-traps" because they apparently capture eel-like nematodes in the wild. While protozoans may be caught by *Genlisea* (although this was not observed), my observations suggest the multicellular, massive nematodes such as those found in the soil where *Genlisea* were growing would be a more significant prey (Figure 2).

It should be noted that we did not see dead nematodes in the *Genlisea* traps. I do not think this is contrary to my hypothesis that they are being captured and digested, because dead nematodes are extremely difficult to detect in the complex internal structures in *Genlisea* traps—only by moving and waving are the nematodes easily seen. However, it cannot be ruled out that the nematodes may be living

Figure 1: A "laboratory for microscopy" on the top of Roraima, in the location *El Hotel II*. The author making sections of *Genlisea* traps. Photograph by Ines Esterkova.

Figure 2: Nematodes observed within a soil sample in Roraima, in the habitat of *Genlisea roraimensis*. Photographs by the author.

in the *Genlisea* traps, contributing to a food web by digesting other captured prey items. (Since the nematodes are not likely to be removing the prey items from the *Genlisea* traps, it would not seem appropriate to refer to them as kleptoparasites.)

It is interesting to note that Barthlott *et al.* (1998) do mention that field observations in Africa did detect the presence of more than nine species of protozoa in *Genlisea stapfii*. It is very possible that the different species of *Genlisea* have different preferences in prey, and this may be a fruitful avenue for future field research (Studnicka, 1996). It is even possible that protozoans caught are of little nutritive value to the plants, or may even act as kleptoparasites.

Heliamphora nutans as a Deadly Habitat for Mosquitoes

It is well known that each *Heliamphora* pitcher bears a small, usually cup-like lid appendage. This appendage has extrafloral nectaries that produce sweet nectar that has been observed to attract ants and other foraging insects (Lloyd, 1942; Bütschi, 1989; Baumgartl, 1993). During our trip on Mt. Roraima we examined the contents of *H. nutans* traps with surprising results. Our observations showed that the majority of the organic material trapped in *Heliamphora* pitchers consisted not of animal prey, but rather of decaying plant leaves that had fallen into the traps. This humus supports a pitcher-bound ecosystem of life forms—in all the pitchers we examined, we found living aquatic mosquito larvae. (Occasionally we found dead mature mosquitoes.) No other insects were found in the pitchers.

So it seems that female mosquitoes successfully visit *Heliamphora nutans* pitchers to lay their eggs, and survive to escape. Furthermore, the eggs hatch and the larvae develop nicely in the pitchers. They are part of a vigorous ecosystem which transforms the dead leaves into assimilable nutrients for the plant. Is *Heliamphora nutans* carnivorous at all? I think so. First, prey are attracted by the hood appendage. Second, I think that at least a fraction of the mosquito larvae may never successfully leave the pitcher upon maturity. For when the larvae pupate and metamorphize into adults, the newly emerged mature mosquitoes may not all successfully escape the narrow traps, especially since it would be their first-ever attempts at flight. The slightest aerobatic mistake would result in their plunging back into the pitcher fluid to be digested, and could account for the dead mosquitoes we found in the trap (although we are not positive the mosquito larvae and the dead mature mosquitoes found are the same species).

Apparently, the *Heliamphora* pitchers are complicated ecosystems that include detritus, algae, microorganisms, and mosquitoes. This special adaptation lets *Heliamphora nutans* thrive in an otherwise hostile environment.

Utricularia and *Brocchinia* Relationships

It is often observed in the literature that *Utricularia humboldtii* on Auyántepui and Cerro Neblina grows in the fluid filled basins of bromeliads, such as *Brocchinia* spp. (Bütschi, 1989; Rivadavia, 1999; Taylor, 1989). I have made field observations that suggest that, at least in the area of Mt. Roraima where *Utricularia humboldtii* thrives and grows to nearly giant proportions, bromeliad urns may be at most a secondary habitat for the bladderwort.

Brocchinia species are widespread and frequent in the areas near Mt. Roraima. A large population of *B. tatei* grows in the cloud forest at the mountain's base (Figure 3), while at least several more specimens occur on the top of the plateau itself. Below the cloud forest, in the margins of the Gran Sabana, the wet meadows contain many more *Brocchinia* plants (*B. reducta* or *B. hechtioides*—the exact identification is not yet certain). However, even though *Utricularia* are found in all these habitats, we found no cases of *U. humboldtii* living in the *Brocchinia* urns!

Figure 3: *Brocchinia tatei* rosettes about 1 m in diameter are numerous in the cloud forest in the basal cone of Roraima. Nevertheless, no *Utricularia humboldtii* grows there. Photograph by the author.

Figure 4: Dense structure of the grassland community of grasses, including *Rhynchospora* species (Cyperaceae). Note that the leaves of the *U. humboldtii* are up to 114 mm wide, a sign of optimal growing conditions. Photograph by the author.

It has been suggested that the long, arching aerial stolons that *U. humboldtii* produces enable the plant to navigate from one bromeliad urn to another (Taylor, 1989) or other parts of the same bromeliad urn (Rivadavia, 2001). However, in the apparent absence of a close relationship between *U. humboldtii* and bromeliads in the area of Mt. Roraima, I propose an alternate purpose for these stolons—I believe they simply let the plant hop from one suitable pool of water to another. This interpretation is particularly supported by the structural nature of the native grassland tussock communities. These tussock communities appear to be ideal for *U. humboldtii*—I have seen huge colonies of plants, growing to comparatively giant size, i.e. leaves up to 11.4 cm (4.5 inches) wide. They are dominated by dense bunches of plants (including *Rhynchospora* spp., *Orectanthe* spp.), as well as *Phragmipedium* spp (Figure 4). The dense growths of these plants break the wetland into a patchy matrix of pools, and prevent *Utricularia* from spreading from one pool to the next. Only by producing the tall aerial stolons, which arch over the tussocks of interfering vegetation, can the *Utricularia* seek suitable new habitats.

This case is an interesting contrast with another bladderwort-bromeliad pairing. I studied *Utricularia reniformis* and the bromeliad *Vriesea atra* in Serra dos Órgãos, Brazil, in 2000. In this situation, a close relationship between the two species was plainly visible. But while the advantage to the *Utricularia* was obvious, it is not clear if the *Vriesea* benefited from the relationship.

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I would like to thank Barry Rice for writing this paper from my notes.

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Literature Reviews

Bradford, J.C. & Barnes, R.W. 2001, Phylogenetics and Classification of Cunoniaceae (Oxalidales) Using Chloroplast DNA Sequences and Morphology. Systematic Botany 26: 354-385.

This paper is interesting because it proposes a "new" hypothesis on the closest relative of *Cephalotus*, the systematic position of which is still somewhat enigmatic. Earlier studies have assigned Cephalotaceae to the order Oxalidales (Cunoniales) without any convincing affiliation. In this study the ordinal position is confirmed, and sequence homology comparisons of the trnL-trnF-region of the chloroplast genome (rbcL homology comparisons being inconclusive) suggest a sister relationship between Cephalotaceae and Brunelliaceae, the clade with these two being in turn sister to Cunoniaceae. The similar floral morphology of these families has already been

pointed out by Engler (Brunelliaceae, in A. Engler & K. Prantl: Die natürlichen Pflanzenfamilien, Nachtrag zu Tl. III: 184, Engelmann, Leipzig 1897). (JS)

Conran, J.G., Houben, A. & Lowrie, A. 2002, Chromosome numbers in Byblidaceae. Australian Journal of Botany, 50: 583-586.

Byblis aquatica: 2n = 16, *B. gigantea*: 2n = 18, *B. cf. gigantea*: 2n = 18, *B. filifolia*: 2n = 16, *B. liniflora*: 2n = 32 , *B. rorida*: 2n = 16. (JS)

Schuiteman, A. & de Vogel, E.F. 2002, *Nepenthes vogelii* (Nepenthaceae): A New Species from Sarawak. Blumea 47: 537-540.

The new species described is related to what is commonly called *N. fusca* from north Borneo but differs in its smaller dimensions, broader, flat lid without appendages, and 1-flowered pedicels. In some respects it approaches *N. faizaliana* but this name is not mentioned at all in the paper. (JS)

Zamudio Ruiz, S. 2001, Una especie nueva notable de *Pinguicula* (Lentibulariaceae) de los estados de Queretaro y San Luis Potosi, Mexico. Bol. Soc. Bot. Mexico 68: 85-88.

Pinguicula calderoniae is described as new. With its long and narrow summer leaves it resembles *P. gypsicola* but the long corolla tube distinguishes the two species and places *P. calderoniae* in section *Longitubus* that so far did not contain any species with long and narrow leaves. (JS)

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Pinguicula 'Tina', A. Slack, Insect-Eating Plants & How to Grow Them: 113 (1986), 15 Oct.

Book Review

Hoogenstrijd, Gert. 2002. Carniflora: Vleesetende planten werking, soorten, verzorging. Approximately 90 color photographs, 50 pages. Paperback (magazine format), 21 × 30 cm (8 × 12 in), cost is 8 Euros (approximately $US8.50) Ordering information is at the end of this review.

Reviewed by Barry Rice

Gert Hoogenstrijd is well known to carnivorous plant growers as being one of those fellows who so often explores parts of the world filled with carnivorous plants, while also being a respected carnivorous plant horticulturist. He recently wrote this magazine-style book about carnivorous plants—a very nice production you should consider purchasing for your collection.

Each page is formatted into two columns. One column contains text in Dutch, the other column contains text in English. The information is of the usual format; each genus of carnivorous plant is mentioned, its mode of carnivory is discussed, and cultivation and conservation issues are described. So while this would be an entirely suitable book for a person first learning about carnivorous plants, anyone with more than two or three books in their carnivorous plant library will learn little new.

However, as with most popular books on carnivorous plants, the photographs contribute a large amount to the value of this book, and make it a worthwhile purchase. The illustrations are very well reproduced and of high quality. Many photographers contributed to the production of this book. (Some of my own photographs appear in this book, but I do not benefit financially from sales of the book.) Illustrations by Art Vogel and Bruce Salmon are particularly luminous. A photograph of *Catopsis berteroniana* plants festooned on a nearly leafless tree on Auyan Tepui (p.23) is the finest I have ever seen of this species.

Referring to this as a magazine is not quite fair, incidentally. The cover is much stronger than usual magazine stock, and the pages have a high clay content—they are similar to the pages of Carnivorous Plant Newsletter in texture. The typography is clean and easy to read.

This book can be purchased from a number of carnivorous plants societies, or from the web site http:// www.extreme-plants.nl, where purchases via major credit cards are possible. You may also contact the author for purchasing details: carnivora2002@hotmail.com. Buy it—you will not be disappointed.

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